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METHODS

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# A low-cost, durable, submersible light trap and customisable LED design for pelagic deployment and capture of fish parasite *Salmincola* sp. copepodids

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## Abstract

Documenting species invasions and assessments of ecological changes depend on detection. Here, we present a simple design of a plankton light trap with specific wavelength LEDs and modifications. We used PVC pipe to create standardised small, rigid, low-cost traps that can be deployed in lentic habitats. With a cost of under $30 US each, including lights and rechargeable batteries, our traps are affordable without the need for disposable chemical lights. These small traps rely on a vacuum to retain contents upon retrieval, eliminating complicated closing mechanisms and allowing bottom entry. Our design includes submersible LED lights that can withstand pressures of at least 5 atm. We expect that the included instructions for underwater light construction and rubber weights using sand may be broadly applicable. However, we designed and field-tested our traps focusing on the detection and capture of the infective copepodid lifestage of a freshwater parasitic copepod, *Salmincola californiensis*. This lifestage had previously only been observed by rearing in a laboratory setting and is of concern due to continued spread outside of its native range and detrimental impacts on salmonids, especially in freshwater reservoirs. We used a 445–450 nm wavelength LED light for capturing *Salmincola* copepodids, but the light design can be modified to any readily available LED and heat sink to attract other target organisms. In our case, the overall affordability of the trap and components allowed for the extensive trapping needed to capture and map the occurrence of rarely-observed species and lifestages, such as the copepodids of *S. californiensis*. In general, increasing the number of traps that can be deployed within or across sites can aid in the spatial comparisons of plankton distributions needed in studies of ecology and species life histories. Light traps may aid in the detection of introduced zooplankton, such as *S. californiensis*, outside of their native range and associated plankton community changes.

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**Keywords**
lentic, parasitic copepod, pressure, reservoir, *Salmincola californiensis*, salmon, zooplankton

## Introduction

Zooplankton are integral to the ecology of aquatic habitats (Lehman 1988), including free-living holoplankton species as well as juvenile life-stages of invertebrates and vertebrates that transition to non-planktonic adult forms. Introductions of both free-living and parasitic zooplankton can have important ecological implications (Yan et al. 2002; Havel and Shurin 2004; Duggan and Pullan 2017). In many cases, zooplankton lifestages contain critical transitions such as host-finding (Heuch et al. 2007) or settling (Freckelton et al. 2017) that are often poorly understood outside of laboratory settings because of the difficulty of field collection. For species of concern, collecting infectious and larval stages from the wild may be important in understanding population dynamics and detecting novel invasions (Suárez-Morales and Mercado-Salas 2013). However, such collections can be challenging in environments that are difficult to sample by tows and when taxa have strong swimming abilities or are otherwise under-represented when using traditional netting collection methods (De Bernardi 1984; Porter et al. 2008).

Many plankton species, especially those with strong swimming abilities, may be phototactic in order to regulate their position in a water column (Martynova and Gordeeva 2010). This can reduce predation risk while maximising foraging opportunities for planktonic consumers that exhibit diel vertical migration, with light acting as an external cue and providing directional orientation (Forward 1988). For organisms exhibiting positive phototaxis, light traps are a logical method of collection (McLeod and Costello 2017). Light trap designs vary widely, but include a light source and an opening that narrows towards the interior of the trap (McLeod and Costello 2017). Often, traps have horizontal entrances (Meekan et al. 2001; Fisher and Bellwood 2002; Hernandez and Shaw 2003) with collection into a mesh cup or basket at the bottom of the trap. Traps are often mesh-sided; however, mesh nets may experience problems with clogging during phytoplankton blooms (Jones 1971) and by non-target animals attempting to enter the trap (McLeod and Costello 2017). Traditional light traps are often expensive and require construction by a machine shop (Floyd et al. 1984). Recent efforts towards inexpensive light traps include modifications of existing water samplers or nets and the use of disposable light sticks (Kehayias 2006; Kehayias et al. 2008), as well as larger designs that incorporate inexpensive materials with a modular light box and fluorescent lighting (Watson et al. 2002). Light traps can be quite large, up to one metre in height (McLeod and Costello 2017), though smaller traps may be as or more effective (Meekan et al. 2001). Designs for small, simple, inexpensive traps and light source customisation to collect phototactic plankton are lacking, especially considering the recent broad availability of selective wavelength LED components.

Light traps may capture species that are absent in other sampling efforts (Jones 1971). For example, *Salmincola* sp. are freshwater Lernaeopodid copepods, known as natural parasites of Brook Trout and other salmonids (Fasten 1912). However, these copepods have been recently recognised as a problem in regions where they have likely been introduced along with host fishes (Kamerath et al. 2009) and in altered systems including reservoirs (Hargis et al. 2014; Monzyk et al. 2015; Lepak et al. 2021). *Salmincola californiensis* is of particular concern because of high infection prevalence and parasite burden on threatened Chinook Salmon (*Oncorhynchus tshawytscha*) and associated impairment of the fish in Oregon, USA (Herron et al. 2018; Neal et al. 2021). Yet, copepodids (the infectious, free-swimming lifestage) of this species were not detected after four years of extensive tow netting in multiple upper reservoirs of the Willamette Basin (Murphy et al. 2019a) where *Salmincola* sp. adults were common on salmon (Monzyk et al. 2015). Unlike sea lice (family Caligidae), *Salmincola* sp. (commonly known as gill maggots) permanently anchor onto their host during development. Their development includes a non-swimming nauplius, a motile copepodid and attached chalimus and adult stages and they are restricted to freshwaters throughout all lifestages (Kabata and Cousens 1973; Murphy et al. 2020). Although adult female *Salmincola* sp. are easily observed on parasitised fish, the infectious copepodid stage has not been captured in the wild. Rather, this stage was described in the laboratory through hatching egg sacs from adult females (Kabata and Cousens 1973). Capturing copepodids could provide early detection where sensitive fish are rarely handled, offer insight into the conditions that are exacerbating infection rates and may eventually lead to methods for management and control. There remain challenges to capturing and quantifying species exhibiting primarily vertical (not horizontal) movements, such as copepodids of the parasitic genus *Salmincola* (Poulin et al. 1990) and to producing a sufficient number of traps to detect their presence and distribution (Doherty 1987), assuming they may be patchy in space and/or time (Herron et al. 2018).

To figure out how we might capture *S. californiensis* copepodids, we conducted extensive laboratory observations and revised the literature about sampling for infectious stages in other groups of parasitic copepods. We noted, as had previous studies for related species, that *S. californiensis* copepodids were negatively buoyant and frequently rested on the bottom of laboratory tanks outside of active bouts of swimming (Poulin et al. 1990). Based on the findings of Penston et al. (2004, 2008) for sea lice, we initially focused on mid-water tows and benthic sleds for copepodid capture. However, such sampling failed to yield copepodids from reservoirs where high levels of infection were documented (Monzyk et al. 2015). After noting positive phototaxis exhibited in the lab and the design by Novales Flamarique et al. (2009), we focused on the design and construction of customised light traps as we did not find replicable published designs that would allow capture from below (but see Tranter et al. 1981). Generally, leaving openings at the bottoms of traps would allow water (and captured material) to flow out of traps during retrieval unless there

was a closing mechanism. Noting the limited information available on the customisation of light sources, on designs for traps with bottom openings (important for captures during the process of vertical migration) and on designs minimising mesh (important for species that are easily damaged or difficult to remove and to avoid clogging during phytoplankton blooms), we designed a novel small, robust trap with customisable lighting that can be run in vertical transects. A non-mesh trap was important because of the presence of structures and debris (e.g. stumps) that could damage mesh traps, as well as phytoplankton blooms that cause clogging of mesh cod ends. Designs using acrylic that fit these criteria were cost prohibitive for the extensive trapping we anticipated for this difficult-to-capture infective lifestage of the parasitic copepod. Finally, the commercialisation of designs for aquaculture has limited the information available to guide light trap construction. Here, we describe how we built, set and collected samples from our low-cost, robust light traps that can be deployed in series from the water surface to depths of at least 60 m and that can be outfitted with selective wavelength LEDs, based on target species. We also provide field test data from three reservoirs in the upper Willamette River Basin in Oregon, USA, to demonstrate the efficacy of these traps.

## Methods

We captured *S. californiensis* copepodids from the wild (Cougar Reservoir, OR, USA, November and December 2018) using our novel light traps. These first captures of *Salmincola* sp. relied on prototype traps constructed using clear, glass funnels set into the bases of 5-gallon (19 l) buckets with sealed lids. Although successful, these traps were large and easily damaged. During associated field trials, many of the underwater lights we tried were ineffective, not modifiable and cost prohibitive. Our study sites are used for human recreational purposes and valuable equipment is more likely to be disturbed or removed. Thus, we explored PVC-based traps to focus on improving portability and durability while maintaining a cost that would be reasonable to deploy at a large scale and that could accommodate loss. The final trap design is based on an 18 cm length of 4-inch (10.16 cm) diameter Schedule 40 PVC pipe that is easy to source in the plumbing section of local hardware stores (Suppl. material 1: Table S1). We were unable to reproduce the closing mechanism in the trap described by Novales Flamarique et al. (2009), even after contact with the authors, so we focused on a simple design that would create a vacuum, thus retaining contents upon retrieval. Lights were placed in water-tight jam jars and either secured to the top lid or secured at the top of the trap by having their latch seated in a cut-out at the top of the pipe (Fig. 1). The light beam was directed downwards through a glass funnel placed in the bottom lid. We designed the traps to use rechargeable AA batteries because they are easy to source and minimise disposable waste after intense use.

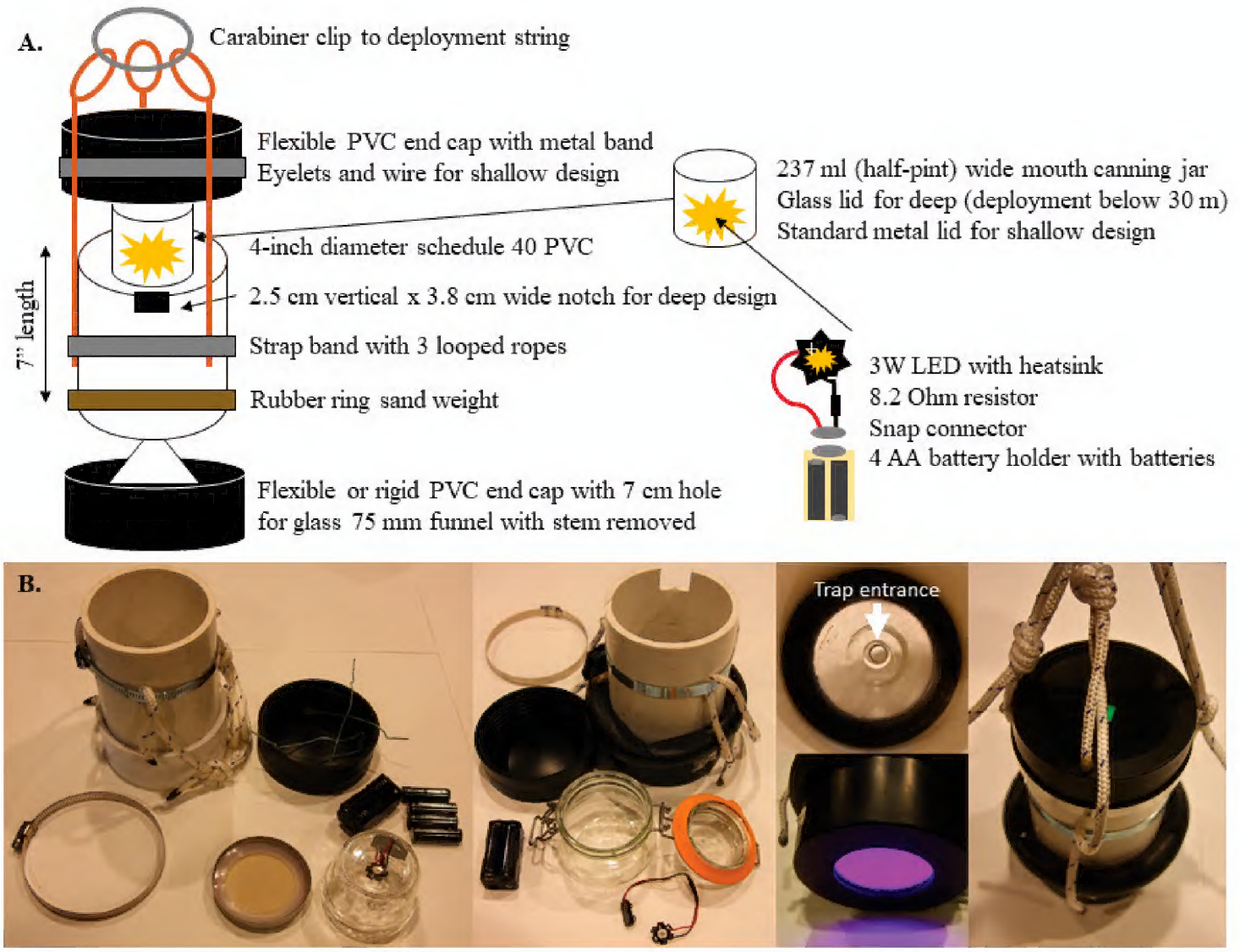


**Figure 1.** Trap design **A** trap schema and component placement. Light is placed under the lid facing down towards the open inward-facing funnel at the bottom (entrance 8 mm at narrowest point, 75 mm diameter at the widest) **B** views of traps including standard (left), deep (centre) and funnel, light colour and deployment loops (right). The rubber weight is used on both trap configurations to ensure vertical orientation, but is pictured on the deep trap here, for reference.

## Trap assembly

### Simple solid selective wavelength LED

We constructed our lights using LEDs that came already affixed to a heatsink. This meant that connections were simple solders of the positive and negative connector wires from the battery snap connectors to the heatsink. We spliced a resistor into the negative wire, although it could be spliced into either wire, and covered the resistor and associated solder connections with heat shrink (Fig. 1). The lights were stored in their respective jars and snapped to the battery holder during deployment. We chose a resistor that would provide more than 48 hours of relatively consistent bright light, based on the battery discharge curve for the rechargeable batteries we used.

## Optional microcontroller and light array

Our initial trials in the laboratory used lights of differing wavelengths and a microcontroller to adjust intensity and mimic shadows. We describe that version here, although we did not complete field testing using it because solid lights were more effective for our target species during lab comparisons. These instructions should aid in the design of more specialised lights and the code (Supplement) can be adjusted to control individual light parameters. Prototype photos, firmware code and a shopping list for the microcontroller array can be found in Suppl. material 1: Fig. S2.

## Trap body (PVC cutting, optional notch and securing lines)

For the trap bodies, we cut PVC pipe into ~ 18 cm long pieces using a mitre saw. This was all that was necessary for traps to be deployed in shallow water (< 30 m). For the traps to be used in deep water (≥ 30 m), we cut a 2.5 cm deep by ~ 4 cm wide notch into the top perimeter of those trap bodies. We marked the opening and cut downwards using a hand-held jig-saw. Channel lock pliers were used to bend the newly-created tab of PVC inwards and snap it off. Notches allowed seating of the rear hinges of the snap closures of glass-lidded jars; we wedged the front latch of the closure fully into the trap body when traps were set. Care was taken not to cut the notch too deeply, as the flexible PVC top lid must seal fully below the notch. Further, we prepared traps for deployment by securing three ~ 55 cm lengths of rope to the trap body with a worm gear clamp. We tied a triple sliding hitch in each length of rope so that fine adjustments to vertical balance could be easily made in the field.

## Lower lid preparation

When flexible PVC lids were used, they were placed on the PVC pipe to stretch prior to construction to minimise issues during assembly and deployment. Lower lids varied in material, but we took care to minimise the edge spaces where trap contents could become lodged and filled any remaining gaps with silicon for the rigid PVC or ABS lids. This extra care was not necessary with flexible PVC lids. Regardless of bottom lid material, we used a drill press with a 7 cm hole saw to make a perforation in the centre of the lid. We attached the lid to the PVC pipe by either hammering it in place (rigid) or by placing it on and securing it with a worm-gear hose clamp tightened with a screwdriver (flexible PVC).

Funnels were prepared by cutting the stem off (before the opening widened) using a hand-held metal file (scoring the stem on all sides). We then lined the large end of the funnel opening with silicone (for adhesion) and firmly placed the funnel inside on the lower lid of the trap to cover the hole in the lid and ensuring a complete seal around the funnel perimeter. We left the trap open and undisturbed for 24 hours to allow the silicone to cure and the funnel to be fully secured.

## Top lid preparation

Top lids were always the type made from flexible PVC. The top lid was used without modification for traps for deep deployment that used glass snap-top jars, because the jars were held at the top of the trap by the notch in the PVC for their closing mechanism. Standard, metal-top jam jars used for traps deployed in shallow water needed to be secured to the top lid. For this, we used four small eyelet screws placed evenly spaced just inside of the ring where the PVC tube is seated when the cap is on. We threaded one wire piece through each screw and secured the wire with pliers. We then bent the wires around each other by hand when loading the light jars during deployment

## Weights

We cut used bicycle inner tubes into sections of approximately 55 cm, discarding the stem valve and pieces with large holes. We sealed one open end of the tube by folding it over and securing it with duct tape. Then we used a funnel to fill the tube with sand. We left ~ 8 cm of the tube unfilled and looped the tube around a length of ~ 10 cm diameter PVC so that the open, unfilled section overlapped the sealed end. We then wrapped the two ends together with tape to seal and secure the weight in a circular form of ~ 12.5 cm diameter. The weights stretched, so there was flexibility in sizing. Bike tube diameters also varied, but weights were only used to keep traps orientated vertically, so exact weighting was not necessary.

## Deployment line features

To allow setting of multiple traps along a vertical transect, we prepared loops at pre-measured distances along a deployment line of a measured length (we made loops at 1 m, 2.5 m, 5 m, 10 m, 15 m and 20 m along a 25 m line) that was also looped at both ends. Loops at the top and bottom of each line were used for attaching an anchor and a buoy. We ensured that butterfly loops along the line were sized to allow the weighted traps to be positioned next to the line without being forced at an angle (e.g. > 9 cm). Anchors were made by adding water to concrete mix and pouring the concrete into ~ 15 cm lengths of concrete form tube (placed on disposable plastic or cardboard). We placed one eyebolt into each concrete anchor, ensuring the bolt was in place and jiggling the mould after placement to settle the concrete around the bolt. Anchors were left to dry for approximately one week, though curing was weather dependent. Additional lines were prepared in pre-measured spools to link deployment lines to buoys when waterbody depths were greater than the length of the deployment lines.

## Trap deployment

We placed a light (facing down) in each jar and secured it into position with a small piece of tape. Then we put batteries into holders and snapped the holders to the light

contacts. Lights were placed in either jars sealed with a silicone ring, metal lid and metal band for shallow deployment (< 30 m) or snap-top jars with glass lids and rubber gaskets for deep deployment (≥ 30 m). The glass-lidded jars were needed for deep deployment because metal jar lids crush from the pressure at depths of more than 30 m. Note: batteries should not be added early to unclipped battery holders, especially in metal-lidded jars, to avoid a risk of shorting.

Jars were fixed into position in the traps either by twisting the wires attached to the lid (shallow) or by being seated into the notch cut into the trap body (deep). We filled a tote with filtered water that we used to fill the traps prior to deployment. After filling each trap with water by submerging it in the tote, we secured the top lid to each trap while submerged and used a screwdriver to tighten the associated clamp, making sure that the clamp held the lid tightly to the trap body with a complete seal. We then clipped each trap to a loop in the deployment string as we lowered the string and associated anchor into the water.

## Trap collection

At each vertical transect, traps were collected sequentially from surface to bottom and placed into white primary wash basins (11.4 l) upon retrieval. These were labelled with the depth for each trap. Once trap lids were loosened and the vacuum was broken, contents flowed into the small primary basin. Wash bottles were used to thoroughly clean out each trap and wash any additional organisms into the wash basin. The basin contents were then sieved using a 106 μm test sieve and washed through a funnel into a collection bottle (250 ml HDPE Nalgene). Primary basins found to contain juvenile fish were nested inside of larger (17 l) secondary basins containing ice for at least 20 min to anaesthetise the fish. All samples were preserved using 95% ethanol with a final concentration of at least 75%.

## Field assessment

We set our light traps monthly from June through to December 2019 at three reservoirs (Cougar, Fall Creek and Lookout Point) in the Upper Willamette Basin, Oregon, USA. From June through to September, we set five vertical strings at each reservoir (Fig. 2) and, on each string, we typically set traps at distances of 1, 2.5, 5, 10, 15 and 20 m from the bottom of the reservoir. Fewer traps were set on a string if water depth at a sampling location was less than 20 m. Similarly, from October through to December, we set five strings per reservoir, but on each string, we set traps at 1, 2.5 and 5 m from the bottom and set additional traps every five metres from the bottom to the surface regardless of the depth at the sampling location, thus adding a few more traps per string at the deepest locations. However, because one of the reservoirs (Fall Creek) was drained to the stream bed before sampling in November as part of normal operations for that site (Murphy et al. 2019b) and the reservoir only partially refilled

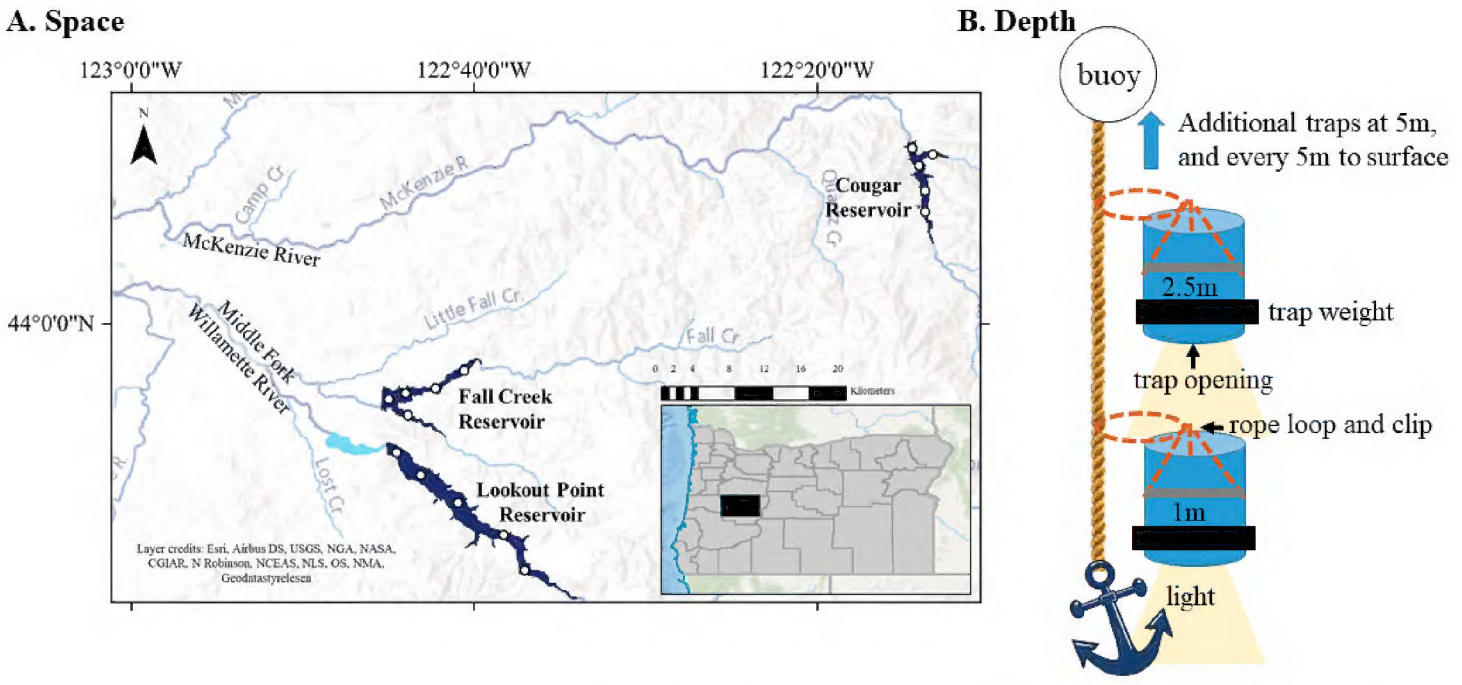


**Figure 2.** Representation of field deployment set-up over **A** space and **B** depth within reservoirs in the Willamette Basin, Oregon. Traps were placed at five sites per reservoir (as indicated with white circles) with traps at 1, 2.5 and 5 m from the benthos continued by traps at 5 m intervals to the surface along a single vertical deployment string held in place by an anchored buoy. Traps were clipped on to pre-tied loops in the deployment rope (represented as dashed orange ovals) to facilitate placement.

in December, fewer strings and traps were deployed at that reservoir in the last two months of sampling.

For our field test, each month, we deployed traps for 48-hour and lights continued to operate throughout deployment. We chose to use 445–450 nm wavelength LED lights in our traps because laboratory trials we conducted showed these were better at attracting *S. californiensis* copepodids than white LEDs or LEDs producing narrow bands of longer or shorter wavelengths (Suppl. material 1: Table S2). The lights we used were set to be always on and not fluctuating in intensity because laboratory trials indicated this was more attractive than when lights fluctuated or operated in series to simulate shadows of passing fish (Suppl. material 1: Table S2). Nevertheless, we provide a microcontroller design as an option for variable light settings in Suppl. material 1:Figs S1, S2 in case that could be useful in other applications and for other species. We added bait to our light traps to increase the probability of capturing target organisms (Burkett et al. 2001). We hoped scent from the addition of salmonid fish tissue would increase the attraction of light traps to the parasitic *S. californiensis* copepodids, but would decrease the attraction of the light traps to non-target organisms that are preyed upon by fish. Inclusion of fish tissue in the traps was accomplished by placing some frozen salmon fin tissue in a teabag and positioning that between the lid of the light jar and the top lid of the trap in each trap. Although we did include bait in the traps for our field test, our laboratory trials indicated that this type of bait did not affect the attraction of light traps to *S. californiensis* copepodids, but it did slightly reduce captures of non-target free-living zooplankton (Suppl. material 1: Table S2).

Field-collected light trap samples were examined in the laboratory as described below. Further, for comparison with light traps results, we also collected monthly vertical tow samples and Van Dorn trap samples at each reservoir (see Murphy et al. 2019a for tow methods and analytical procedures).

## Laboratory methods

Light trap samples were processed in the laboratory to identify and count *S. californiensis* copepodids and other non-target organisms that were also captured. To increase our efficiency at searching for *S. californiensis* copepodids, we washed each sample through a pair of stacked sieves (Fig. 3A). The upper sieve had a 500 µm mesh size which retained mature *Daphnia* spp., large calanoid and cyclopoid copepods and other large organisms (e.g. fish larvae, *Leptodora* sp. and aquatic insects), but allowed *S. californiensis* copeopodids and smaller zooplankton to pass through (Fig. 3B); the lower sieve had a 106 µm mesh size to retain the size fraction of zooplankton that included *S. californiensis* copepodids (Fig. 3C). We washed plankton retained on the mesh of the 106 µm sieve into a Petri dish with 95% ethanol and searched this

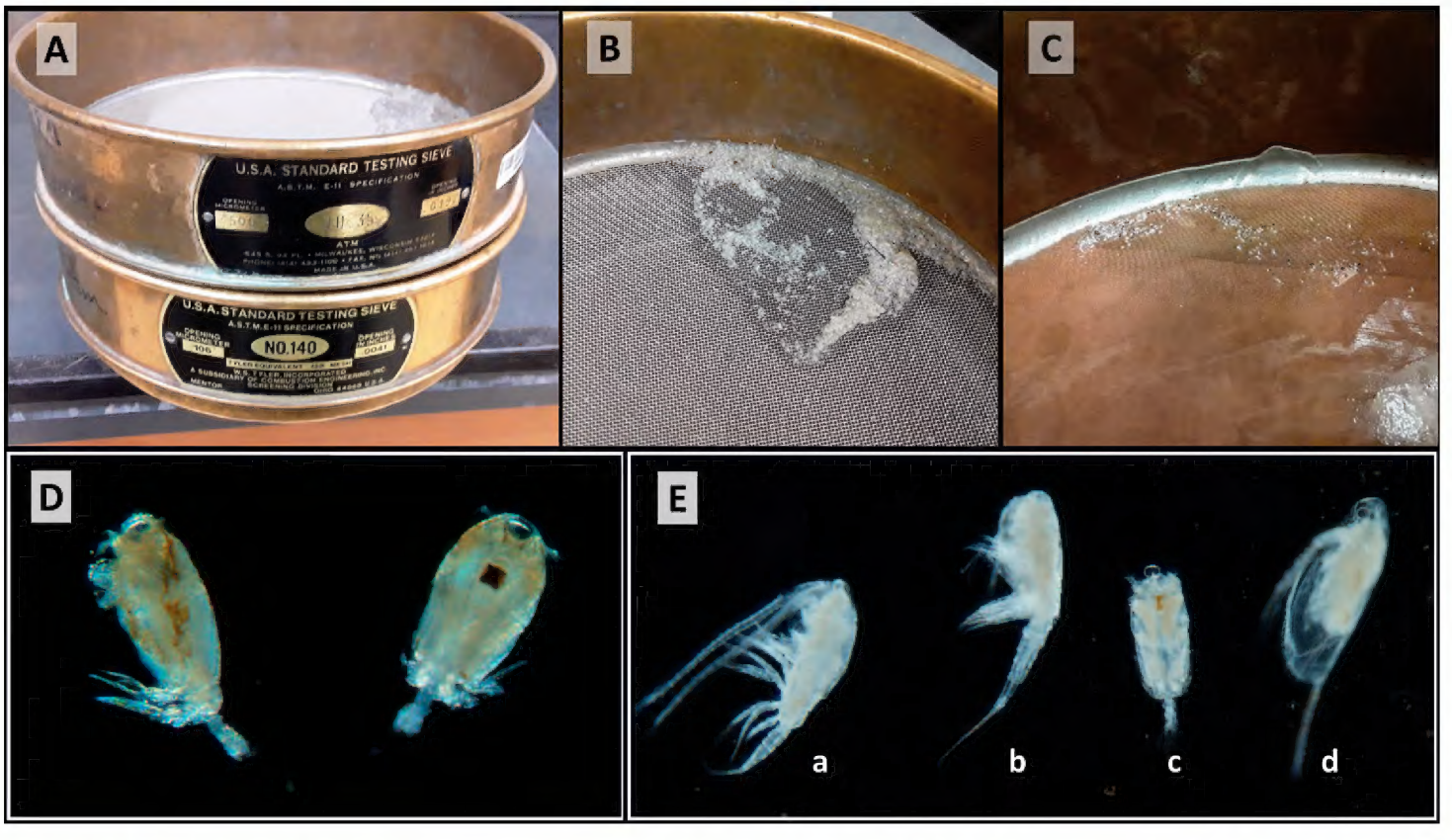


**Figure 3.** Photos of sample processing and characteristics of *S. californiensis* copepods compared to other common zooplankton in light trap samples **A** stacked sieves used in light trap sample processing **B** large zooplankton retained on the 500 µm sieve **C** smaller zooplankton that could include *S. californiensis* copepodids retained on the 106 µm sieve **D** freshly preserved *S. californiensis* copepodids from a laboratory rearing experiment **E** comparison of common zooplankton caught in a light trap and retained on a 106 µm sieve including: a) a calanoid copepod, b) a cyclopoid copepod, c) a *S. californiensis* copepodid and d) a *Daphnia* species.

material thoroughly under a dissecting microscope at 8X-20X magnification. Any *S. californiensis* copepodids found were recorded and removed from the sample.

In practice, distinguishing *S. californiensis* copepodids from other zooplankton was not difficult. We had examined many of these copepodids during laboratory hatching and rearing experiments (Murphy et al. 2020) and there are excellent illustrations and descriptions of the morphology of *S. californiensis* copepodids in Kabata and Cousens (1973). We identified other organisms in the light trap samples using keys and illustrations in Thorp and Rogers (2016). In comparison to free-living copepods, *S. californiensis* copepodids have a different body form including a lack of extended first antennae modified for swimming, a transparent lens-like structure at the anterior end of the body (the terminal plug of the frontal filament used to attach to a host fish) and a pigmented eye spot containing three ocelli in the middle of the dorsal surface of the anterior portion of the body (Fig. 3D, E). The pigment of the eye spot, however, can fade somewhat with prolonged storage in ethanol preservative.

After searching for and removing *S. californiensis* copepodids from each light trap sample, we drained the alcohol from the remaining material in the Petri dish using the 106 μm sieve and rinsed this material and the material retained on the mesh of the 500 μm sieve into a beaker with water; a couple drops of detergent were added to prevent organisms from clumping. We then subsampled these non-target organisms using a Folsom plankton splitter to target 200–400 organisms for representative identification and enumeration.

## Results

We successfully captured *S. californiensis* copepodids using our light traps, but did not detect them in concurrent tow or Van Dorn traps samples (Table 1). In total, we collected 532 light trap samples across all reservoirs and sampling dates and 53 of the traps captured our target organisms. For the traps that captured *S. californiensis* copepodids, the number of copepodids captured ranged from 1 to 169 with a median of 2. Non-target organisms were also captured in our light traps (Suppl. material 1: Table S2). Most of them were cladocerans (primarily *Daphnia* spp.) and calanoid and cyclopoid copepods. Other organisms caught in lower numbers were various aquatic insects, aquatic mites, ostracods, amphipods and larval fish. Captures of all organisms in light traps were highly variable depending on reservoir, collection date and trap depth. Interestingly, *Salmincola* copepodids were captured at shallower and warmer conditions than expected, based on documented copepodid survival (Murphy et al. 2020; Suppl. material 1: Table S2).

## Discussion

Here, we describe light traps that successfully attract and capture *Salmincola* copepodids from lentic systems with the potential to be used to examine patterns of both

spatial and vertical distribution of this species in our study reservoirs. In order to sample for this 'needle in a haystack', we present a robust low-cost trap design that can be widely deployed, including at depths with pressures of 5.9 bar and in environments that could damage mesh-based traps (e.g. reservoirs with large stumps and woody debris). The simple design eliminates the need for complex and expensive closing mechanisms and is constructed from easily-obtained materials. By establishing methods to capture this poorly-understood copepodid life stage, we will be able to gain valuable ecological knowledge about its density and distribution over time and space. This information should help us to understand the current problem of high infection prevalence and intensity and may allow us develop remediation strategies. In addition, the

**Table 1.** Number of light traps that captured *Salmincola californiensis* copepodids in our field trial at three upper Willamette Basin reservoirs. Samples were collected monthly from June through to December 2019. Number of traps set given in parentheses. Note: corresponding tow net samples from 35 m to surface (0.5 m diameter, 64 μm) failed to capture copepodids during the study period.

| Reservoir | June | July | August | September | October | November | December |
|---|---|---|---|---|---|---|---|
| **Cougar** | 0 (25) | 1 (29) | 3 (28) | 10 (28) | 7 (33) | 7 (29) | 8 (28) |
| **Fall Creek** | 0 (23) | 0 (26) | 0 (26) | 8 (28) | 7 (23) | 0* (2*) | 1* (11*) |
| **Lookout Point** | 0 (24) | 1 (27) | 0 (27) | 0 (28) | 0 (33) | 0 (30) | 0 (24) |

* Fall Creek Reservoir was drained prior to sampling in November and only partly refilled when sampling occurred in December leaving less habitat available to sample in those months.

use of our light traps provide monitoring data to prompt ecological discoveries (e.g. use of optimised traps as potential method of control and management of copepodids).

We focused on 48-hour deployments because it increases trap captures and allows for us to set strings at multiple sites during our sampling window. Captures are thus possible throughout the day and night periods. Even so, we would expect most captures to happen at night, when the lights provide a greater difference in illumination from background natural levels. We did not focus on new moon periods, though moonlight could modify capture efficiency (Hickford and Schiel 1999). Light intensity is also likely important and we use bright lights because other species of parasitic copepod are more attracted with increasing intensity (Novales Flamarique et al. 2009).

Inexpensive traps can be important to achieve research goals with limited funding. We expect these traps to be especially useful for rare or poorly-understood species where large-scale deployment may be necessary. They are also ideal for areas with boat traffic where traps may be stolen or damaged. The option to deploy traps at depth may provide greater captures, as other studies have found increased catches near benthic areas (Tranter et al. 1981; Tor et al. 2009). Their small size can make plankton easy to damage by netting and pumping (Beers et al. 1967); damage is less likely with light trapping methods, especially using rigid traps, because taxa are not forced against a net. Inexpensive lights that function below 30 m are likely to be useful in an even broader context, as additional sampling and surveying methods rely on light for attraction or

visibility. While here we use 445–450 nm wavelength LEDs, a wide variety of options are available. Spectral sensitivity for *S. californiensis* at 445–450 nm would not be unexpected given that other freshwater taxa have shown similar maxima (Forward 1988). Longer wavelengths (550 nm) have been documented as the maximum sensitivity for marine parasitic copepodids (Bron 1993) and peak sensitivity to 475–525 nm wavelengths is common for taxa in estuarine, coastal and open ocean habitats, though some taxa may have peak sensitivity as low as 360 nm (Forward 1988).

Future studies could explore the efficiency of these traps, especially for other taxa and the changes in capture composition with alternative LEDs and/or differing LED intensities. Blue and green wavelengths are likely ideal for a broad range of taxa and transmit well in water (McLeod and Costello 2017). Using a microcontroller, while not pursued in our final design, would allow for lights in series, patterns in illumination and the fine tuning of light intensity. Shorter duration deployments could explore day versus night efficiencies and may allow for the use of smaller batteries and possible further downscaling of the design presented here.

Outside of light trapping, we expect that the instructions (Supplement) for underwater light construction and rubber weights may be broadly applicable. Our description of how to assemble LED light sources for use underwater allows for researchers to customise wavelengths and target desired taxa. Low-cost selective wavelength LEDs may also be useful for other underwater applications (e.g. video). Weights that use discarded materials (bike tubes) and sand can aid in balancing equipment underwater where cost or vandalism are of concern. In our case, the overall affordability of the trap and components allows for the extensive trapping needed to capture and map the occurrence of rarely-observed species and lifestages, such as the copepodids of *S. californiensis*. In general, increasing the number of traps that can be deployed within or across sites can aid in the spatial comparisons of plankton distributions needed in studies of ecology and life history. Effective trapping of zooplankton, parasitic and free-living, may be critical to early detection of invasions or novel ecological dynamics.

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## Supplementary material 1

**Supplementary material for a low-cost, durable, submersible light trap and customizable LED design for pelagic deployment and capture of fish parasite *Salmincola* sp. copepodids**

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Data type: Pdf file.

Explanation note: **Table S1.** List of materials used to construct light traps and deployment strings. **Table S2.** Laboratory captures in light trap development. **Table S3.** Additional materials used for microcontroller programmed lights in series. **Table S4.** Examples of plankton captures categorised as Cladocera, Calanoida, Cyclopoida and *Salmincola californiensis* for Fall Creek and Cougar Reservoirs in late October 2019. **Figure S1.** Wiring diagrams. **Figure S2.** Prototype light configuration with Arduino and firmware code (.ino) for varying the light intensity in series. **Manual S1.** Pictographic light trap assembly instructions.

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